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24. Trapping, collaring and monitoring the Lorisinae of Asia (*Loris*, *Nycticebus*) and Perodicticinae (*Arctocebus*, *Perodicticus*) of Africa

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ABSTRACT

We can gain a better understanding of elusive nocturnal mammals through live trapping them to gain vital samples and measurements and through following them in the night with radio or GPS collars. Studies that involve humane live-trapping of a species open the doors into better understanding a species' evolution, diversity, adaptations, and behaviour. We review 11 studies that have attempted to trap lorises and pottos with physical traps, and show that this method yields very low success, and is achievable largely with traps specifically built and designed for the species. Hand trapping, however, was successful in 7 studies, and we make recommendations for this procedure. Once caught, we provide a detailed list of measurements and samples that can be taken, and review 14 studies (both reintroduction and wild) that have used various radio collars to track lorises and pottos. We intend that these recommendations will provide a standard by which lorises and pottos can be compared throughout their range.

Key Words: trapping, radio tracking, radio collaring, behavioural observation, Lorisidae, Lorisinae, Perodicticinae,

1. Introduction

Nocturnal animals are difficult to see and follow, especially in dense rain forest conditions. Nocturnal research is fraught with difficulties not encountered by individuals who study animals in the day, from the need for expensive equipment, constant access to power supplies to run lights, and potential for increased encounters with dangerous wildlife. The main drawback of nocturnal fieldwork is that it is simply more difficult to find and continuously observe an animal at night. Through hard work and perseverance it is possible to obtain ecological data on lorises and pottos in the absence of radio tracking (*e.g*. Nekaris, 2001; Pliosungnoen *et al*., 2010; Das *et al*., 2014). Much more detail can be obtained, however, through capturing, measuring, collaring and monitoring nocturnal primates. The essential nature of radio tracking for the study of the behaviour and ecology of nocturnal primates has been recognised since the 1970s (*e.g*. Charles-Dominique, 1977; Charles-Dominique and Bearder, 1979), and is by most researchers considered a must for thorough research (Sterling *et al*., 2000). Radio tracking studies of lorises and pottos remain limited (Table 25.1). Still, these studies have revealed important information regarding ranging behaviour, selective habitat use, minimum area requirement for a species, effects of habitat fragmentation on behaviour, as well as social organisation. radio tracking allows the researcher to find animals regularly in the forest at night, making efficient use of research time. Once collared, it is possible to monitor daily and seasonal movements of animals, to analyse demographic differences in these movements, to evaluate resource selection, change in home range over time, random versus non-random habitat use, to estimate population densities of animals, and even to estimate vulnerability of extinction (Kenward, 2000; Millspaugh and Marzluff, 2001). In this chapter, we review the methods for trapping and collaring slow lorises and pottos, as well as provide a case study of the importance of red light for observing their behaviour in a humane and productive manner (Box 25a).

1.1 Live trapping of lorises and pottos

In order to collar an animal, one must first be able to catch it. The successful implementation of live-trapping methods has resulted in a better understanding of the behaviour and taxonomy of many once unknown species. Being able to capture animals allows researchers to identify individuals with confidence through marking or microchipping; to measure them and assess age and sex classes; to collect samples to assess health, stress, diet, genetic aspects, *etc*.; to radio collar or GPS tag individuals to calculate home-ranges and infer other behavioural ecology aspects. Many primate species are challenging to trap, with the lorises and pottos being particularly difficult (Nekaris & Bearder, 2011).

As of 2019, we could locate 11 studies that have attempted to catch seven species of loris or potto using some form of trap, only four of which were successful (Table 25.1). With the exception of Wells *et al*. (2004), who accidentally caught slow lorises in small mammal traps, researchers used eight types of traps, with the most successful traps being those designed specifically for lorises and pottos. Despite the 'success' of four of these studies, substantial effort was required to catch only a small number of animals (Table 25.2). Successful captures appear to occur when a high effort is coupled with a long duration. Data for the Central potto imply that at least six months is needed to obtain any results. Except for one adult male potto, it took the majority of pottos six months to start using the traps (compared to sympatric bushbaby species that used the traps within several weeks of being installed) (Pimley, 2003). Still some advice can be obtained from these studies.

Trap type was vital for the success of catching a loris or potto. Commercial traps (*e.g*. Havahart, Sherman, Tomahawk) are designed based on an animal's size and weight. Trap producers do not consider an animal's locomotion, as many animals, including many nocturnal primates, jump, bound or leap into traps and are easily caught (Harcourt & Nash, 1986; Kays, 1999; Radespiel *et al.*, 2003; Schulke & Kappeler, 2003). Lorises and pottos, however, are cautious and hold on to a branch with a back leg, while they stretch their body to get the bait inside the trap (Pimley, 2003). Rarely will they put their entire body in a small trap. Commercial traps for the size and weight of lorises are too short in length when factoring in the loris' stretch. If a larger trap is used, the small body size of a loris will not set off the trigger. Thus, of the four successful studies, three used loris-specific traps. A successful trap designed to catch pottos in Cameroon relied on the animal removing bait from either a transparent Perspex platform with wire mesh traps or a metal hook with wooden traps (Pimley 2003). The action of removing the fruit from the trap served as the trigger to close the trap rather than relying on the animal standing on a platform as a trigger. To reduce stress and increase safety and comfort of the animals, we suggest placing branches/foliage inside and around the trap, and make it with soft materials, like plastic mesh, and not making the interior too large (c.f. Schulze, 2019.). Camouflaging the trap by application of green paint and moss/leaves before being set can also lure animals in whilst deterring predators or human hunters. We also recommend checking the traps on a regular basis to minimise the risk of injury/mortality to trapped animals. Considerations on frequency of trap checking should take account of the local weather (with extremes in hot/cold/wet weather necessitating more frequent trap checking). Care should also be taken to ensure that there are no ant trails near to trap locations, as certain species of ants can eat trapped animals alive.

Bait is key for luring animals into a trap. Lorises and pottos have a diet that ranges from omnivorous, including insects, gum, birds and small mammals, to almost exclusively faunivorous or exudativorous (Charles-Dominique, 1977; Nekaris & Jaywardene, 2003; Pimley & Bearder, 2013; Cabana *et al*., 2017). Only nine of the trapping studies we reviewed detailed the type of bait used. Ripe and smelly banana, sometimes combined with peanut butter, was relatively successful for pottos, which seem to be the only loris with substantial fruit in their diet (Charles-Dominique, 1977; Pimley 2003, 2005a; K. Davey, pers comm.). Wiens (2002) had very small success with banana and more success putting traps around nectar inflorescences, with such traps needing to be manually triggered by a human observer. Efforts to catch the insectivorous slender lorises and angwantibos have not yet yielded any positive results (Charle-Dominique, 1977; Gupta, 2007; Nekaris, this study).

Another aspect considered vital to those studies where trapping success was achieved is an acclimatisation period. A minimum of two weeks is recommended, where the traps are baited, left open but not set, allowing the animals to enter and exit the trap safely. This technique enables the study species to become habituated to the traps thereby enabling recapture of individuals following initial capture (Pimley, 2003).

Location of the trap is also important. Lorises and pottos are characterised as cautious, typically slow movers and are found from heights of 0-30+ m, rarely descending to the ground (Charles-Dominique, 1977; Wiens, 2002; Nekaris & Jaywardene, 2003; Munds *et al.*, 2013). Lorises and pottos tend to favour areas of dense liana and foliage coverage (Charles-Dominique, 1977; Wiens, 2002; Nekaris & Jaywardene, 2003; Pimley *et al*., 2005a), but these areas do not always ensure a safe or secure branch to support the weight of a trap. For Central pottos, the traps were tied on tree branches where possible. Where it was not feasible to climb the tree, they were placed on a platform of thin branches and hoisted up into the canopy at height range 3-5 m. Where possible, the traps were attached securely to branches that were part of the pottos' normal travel route (Pimley *et al*., 2005a & b). Despite unsuccessful trapping for the Philippine slow loris, it is useful to explain the systematic methods attempted. Three areas were selected and prepared for arboreal trapping, based on sightings during nocturnal surveys (Munds *et al.*, 2013). Each location was at least 1 km away from the other trapping spots. At the trapping spots, a 200 m trapping line was created that followed a section of a trail. Ten trees were selected along these trails (30 trees total). One tree was where a loris had been spotted and the remaining nine were spaced at least 10 m apart along either side of the loris tree. A pulley system was set-up that allowed traps to be hoisted to various levels of the canopy. The method for this required the use of a 6-foot tall sling-shot, that projected a line into the tree. The trap was then attached to the line, and an additional line was attached to the bottom of the trap in order to pull it down if the traps were to get stuck. From 9 August- 13 September 2009, all 30 traps were hoisted between 8-30 m and suspended nearby or rested on a branch. Traps were set an hour before dusk (17:30) and checked every six hours (18:00; 00:00; 06:00) by using a torch with a red-filter to check for eye shine or movement. Starting at 06:00 traps were brought down and closed to prevent capture of diurnal animals.

1.2 Live hand catching of lorises and pottos

Catching lorises and pottos by hand has yielded much more success than live trapping, with all seven studies we could locate being successful. The drawback to this approach is that it requires many hours of active searching in an area of reasonable loris density, followed by waiting for an animal to move to a relatively isolated tree. Ideally the animal will be low enough to reach, at which time the branch can be bent, or even snapped off while the animal is still holding on. The animal should then be covered with a soft cloth, or grasped with a snake grip around the neck. We recommend placing the animal as soon as possible into a small, clean, cotton drawstring snake bag for the safety of the animal and the handler. In habitats where trees are very tall and animals rarely come low, hand catching requires climbing of a tree or other substrate – *e.g*. bamboo. Climbing equipment can be used, but by the time it is set up, the animal may have already moved to another tree, thus having expert tree climbers is important in some habitats. Whilst a climber is in a tree, a ground team must be prepared with a tarpaulin in case the animal should fall or drop itself from the branch. The ground team should also guide the climber by watching the movements of the animal, ideally whilst using red head torches. In general a team of about five people are recommended for hand catching, with 2-3 in the tree and the others on the ground (Nekaris, this study; Gupta, 2007).

Hand catching may include a noose, such as used by Charles-Dominique (1977) for *Perodicticus edwardsi* or by luring animals onto a branch, used for *Loris lydekkerianus nordicus* in Sri Lanka and *Nycticebus javanicus* in Java. Noose-catching is favoured by local hunters in Indonesia and Cambodia (Nekaris & Munds, 2010). This method needs to be carried out carefully as to not injure the animal. A forked branch or commercially available snake catcher or 'critter gitter' can be used to immobolise an animal so it can then be retrieved from the tree.

Catching with the hands only, however, has been the single most successful method to capture slender and slow lorises. In Sri Lanka studies of *L. tardigradus tardigradus* and *L. l. nordicus* yielded the catching of 29 animals (12 females, 17 males) and 4 animals (1 female, 3 males) respectively (Nekaris and Jayewardene, 2003; Nekaris and Wijeyartne, 2009). Gupta (2007) caught 22 males and 12 females of *L. l. malabaricus* in South India. Starr and Nekaris (2013) caught 6 female and 7 male *N. pygmaeus* in Cambodia. In Sabah, Stark and Gossens (pers. comm.) caught 7 *N. menagensis*. During a long-term study of *N. javanicus* in Java, between April 2012 and January 2019, researchers caught 89 animals 1-17 times, managing 327 captures. Of these captures, 53% were of females and 47% were of males (Nekaris, this study).

2. Measuring and Collaring

After an animal is caught, immediate efforts should be taken to reduce its stress. The animal should be covered with a cloth or placed in a clean cotton bag so it can hide. If placed in a bag or carrying case, it should be provided with a stick or other implement to which to cling. Care should be taken with any container made of wire mesh, as the mesh can injure the animal's hands or nose. The animal should be transported to a clear space where it can be processed. Gupta (2007) collared animals in a field station, and Pimley (2003) created a clinic in her field station to collar pottos and to collect an array of samples. Starr and Nekaris (2013) initially took *N. pygmaeus* back to camp for collaring, and then realised it was less stressful for that species to be collared *in situ*. Nekaris (2016) created a makeshift measuring area in the forest on a plastic tarpaulin to ensure a clean area where all equipment could be easily organised (Fig. 25.1).

After catching an animal and bringing it to a secure location, the researcher should be ready with a standard set of equipment. We recommend to keep a portable capture kit in a fishing tackle box with multiple compartments. Essential equipment include: a medical kit for loris and human, including an EpiPen in the case of a slow loris venomous bite; radio collar with strong operational signal and a spare, as well as items to secure the radio collar (for example, epoxy resin, shrink wrap rubber coating and a lighter); spring scale (Pesola); sliding electronic callipers; measuring tape; leather gloves for loris handling and latex gloves for sample collection; anaesthesia and its reversal; variety of sample bottles, swabs, and envelopes; microchips and scanner; operational data loggers; wristbands (used for birds) for animals too young to collar; extra pencil and pencil sharpener; camera; spare data sheets; waterproof datasheet holder; plastic bag for rubbish. We recommend carrying the equipment in a water proof bag, and if work is done in the field, to arrange equipment on a clean and dry tarpaulin (Fig. 25.1).

We recommend to take a standard set of measurement data. We use a standard printed data sheet that includes a number of important variables that should be written immediately on the day of capture (Fig. 25.2). It is tempting to think you can fill in the data sheet later, but this procedure often leads to lost and forgotten data. Key data include: the name of the data collector; habitat type; GPS point; age and sex; any injuries or parasites; notable markings; behaviour during capture; records of all photos, videos, sound recordings and samples taken and careful labelling of such samples. In addition, we include a large variety of head and body measurements, as well as detailed descriptions of pelage and facial markings that should be accompanied with diagnostic photos taken alongside a Munsell (or similar) colour standard chart (Fig. 25.1). Our data sheets also include a drawing of a generalised loris so that any special wounds or markings can be drawn for further identification. Measurements are important for several reasons and we highly encourage researchers to adhere to published measurement standards to ensure comparability across studies (Nekaris and Jaffe, 2007;

Poindexter and Nekaris, 2017; Schulze 2019). Measurements can help to determine the age and sex class of the animal; can be used to understand sexual dimorphism, development and growth; can help to assess health, including pregnancy. Equipment we recommend for measuring include: weight taken with a weighing scale (even if an animal is aggressive, it is important to weigh it); sliding callipers for most body measures, with electronic callipers speeding up the process significantly; a soft measuring tape for body length, chest girth and neck girth – ensure that the length of this tape does not change in humidity. Measurements also can help clarify the taxonomic status of lorises and pottos (Ravosa, 1998; Munds and Nekaris, 2010). If the governmental authorities allow, a hair sample with the follicle or other genetic sample (ear punch, blood, faeces), collected and stored properly is invaluable for DNA analysis, both at the species level and to determine information about the mating system of lorises and pottos. A freezer can also be invaluable, with a generator available as a backup to preserve samples. RNA-later solution is also recommended for field conditions.

Most species of loris can be measured without anaesthesia and this is recommended whenever possible. In our experience, slender and slow lorises that have undergone anaesthesia are more likely to fear the researcher and less likely to become habituated, are disorientated for many days upon being released, and are more susceptible to predation. For Central pottos, anaesthesia was used without habituation problems. If anaesthesia is used, Zolitol and its reversal are much more humane. It is highly inadvisable to anaesthetise a pregnant/ lactating female; thus a qualified and experienced veterinarian may need to be present to determine these factors. We also advise against catching a mother when she is still carrying her young. We also have found negative effects of catching an animal in its sleep site, in that it will no longer feel safe using that site again. We thus advise capturing at night.

Lorises can be fitted with small radio collars, 5% less than their body weight (Millspaugh and Marzluff, 2001). A leather collar is soft and prone to falling off and may be better in reintroductions or dispersals where animals may move far from the natal range (Kenyon, *et al*., 2014). A cable tie collar is quick to apply, and lasts for a longer period, but care must be taken to ensure the entire length is covered with plastic shrink wrap (Fig. 25.1, 25.2). Some tips to applying the radio collar include: fit the collar over a pinkie finger or a pencil to ensure it is not too loose or too tight; use an old collar to determine the length of a new collar; always ensure that the collar is functioning before you put it on an animal; note the date of purchase of all collars as they begin to lose their battery power upon manufacture. Young animals should not be collared or should be vigilantly monitored if collared as they will grow and can be strangled; thus measurements of adult neck girth are vital to know the age at which a collar can be safely applied. A plastic bird band or beads on the wrist or the collar can be used to mark individuals so that they can be identified from a distance; if two or more animals are moving together, this helps even when they are wearing collars. Reflective bands, however, should not be used as this can increase predation. All collars should be removed by the end of the study; it is unethical to leave an animal with its collar on, and all attempts should be made to recover the collars. Collars can be refurbished and used again at a lower price, so it is also economically advisable. Table 25.1 details various collars used in loris and potto studies, and Figures 25.1 and 25.3 provide examples of the collars.

3. Problems of trapping, conservation & the future

Many researchers do not want to make public a successful trapping regime when the main threat to most lorises and pottos is the pet and medicinal trade (Nijman & Nekaris, 2014).

Additional threats include the rapid loss of habitat and hunting for human consumption (Svensson & Friant, 2014). Results that can benefit multiple studies, but harm the survival of a species, put loris-scientists in a quandary when it comes to sharing ideas openly, and explains with good reason why trap types are often described in vague terms as “loris-specific” and “wire-mesh”. Sharing too much information may not only benefit the researcher who wants to know how to trap a loris or potto, but it also benefits the poacher. Having said this, a reason hand catching is so successful is that it is the method of choice adopted by hunters, and indeed, in four of our studies, we employed former hunters to help hand trap pottos and lorises, having the positive effect of providing an economic alternative to hunting. Thus we can also say that by reporting these results we are not sharing any new information with poachers. Still, in studies where physical traps are used, it is essential that researchers ensure that their traps are taken down after the study period and in areas with hunting problems, it may be necessary to destroy the traps to prevent their inappropriate usage. Working with each other, openly discussing ideas, will help in creating a trapping regime that will produce more positive results while safeguarding the welfare and survival of these elusive primates.

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Table 1: *Studies of lorises and pottos using radio tracking as of May 2019, including specifics of the model and weight of collar, and the topic and country where the research was conducted.*

Species	Collar Brand (weight - g)	Duration (days)	Topic	Country	Reference
Nycticebus coucang	Biotrack TW-3 (12)	137	Home range, social organisation	Peninsular Malaysia	Wiens, 2002
N. javanicus	Biotrack TW-3 (17)	2,920	Home range, social organisation, territoriality, conservation	W. Java, Indonesia	Nekaris, 2016
N. javanicus	Biotrack TW-3 (17)	16-409	Reintroduction	W. Java, Indonesia	Moore et al., 2014
N. menagensis	Biotrack TW-3 (17)	--	Home range	Sabah, Malaysia	Stark & Gossens, pers. comm.
N. borneanus	Biotrack TW-3 (17)	270	Reintroduction	Borneo, Indonesia	Anirudh, et al., this volume
N. pygmaeus	Sirtrack, (6)	210	Home range, effects of fragmentation & hunting	Cambodia	Starr and Nekaris, 2013
N. pygmaeus	Holohil PD-2C (4)	134	Reintroduction	Northern Vietnam	Streicher, 2004
N. pygmaeus	Holohil PD-2C (4)	300	Reintroduction	Northern Vietnam	Poindexter et al., 2017
N. pygmaeus	Holohil PD-2C (4)	73	Reintroduction	Southern Vietnam	Kenyon et al., 2014
Loris tardigradus	Biotrack TW-4 (6)	339	Home range, social organisation, fragmentation	Southern Sri Lanka	Nekaris & Wijeyeratne 2009
L. lydekkerianus malabaricus	Holohil PD-2C (4)	300	Home range, social organisation	Tamil Nadu, India	Gupta, 2007
Perodicticus edwardsi	Biotrack TW-3 (12)	134	Home range, social organisation	Cameroon	Pimley, et al. 2005b

Table 2: Efforts, methods and duration of attempts to use physical traps to capture lorises and pottos as of May 2019

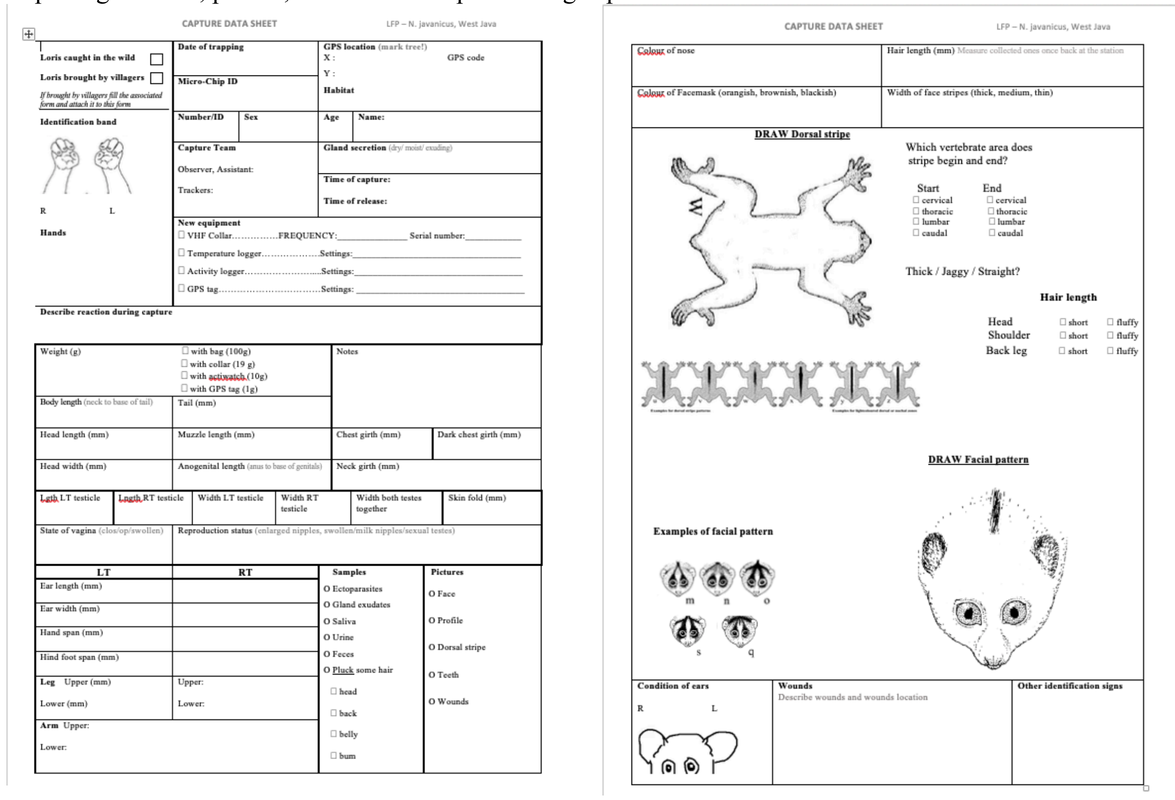
Species	Effort	Trap Type	Average height (m)	Primary baits	Duration	Successful capture (Y/N)	Location	Study
N. menagensis	451	3 different loris specific	15	Jack & palm fruits, figs bananas,	26 nights	N	Lower Kinabatangan, Floodplain, Sabah, Malaysia, Borneo	Munds, this study
N. menagensis	46000	Wire mesh	Ground & 13.5	Banana	6 months	Y	Kinabalu National Park, Sabah, Malaysia, Borneo	Wells et al., 2004[1]
N. coucang	48000	Wire mesh & loris specific	NA-arboreal	Banana, bertram palm	800 nights	Y	Manjung District, Perak, Malaysia	Wiens, 2002
N. bengalensis	33	Wire mesh	8	Banana	3 nights	N	Khao Ang Runai Wildlife Sanctuary, Thailand	Pliosungnoen et al., 2010
P. edwardsi	NA	Noose traps & wire mesh	NA-arboreal	Banana	42 months	Y	Makokou, Gabon	Charles-Dominique, 1977[2]
P. edwardsi	23400	Potto specific wire mesh	3-5	Banana and avocado	780 nights	Y	Mount Kupe, Cameroon	Pimley et al., 2005a[3]
P. potto	602	Potto specific wire mesh	15	Banana	154 nights	N	Kakamega Forest, Kenya	Davey, pers. comm.
L. t. tardigradus	25	Chardonneret box traps	2-3	Banana & treacle honey	5 nights	N	Masmullah Proposed Forest Reserve, Sri Lanka	Nekaris, this study
L. t. tardigradus	300	Chardonneret box (10) & Tomahawk (10)	1.5-3	Banana, treacle honey, cat food, crickets, cockroaches	15 nights	N	Masmullah Proposed Forest Reserve, Sri Lanka	Nekaris and Pimley, this study
L. t. tardigradus	660	Tomahawk, locally made	2.6	Banana & treacle, cat food, crickets	30 nights	N	Bangamukande Estate, Galle District, Sri Lanka	Nekaris, this study

L. lydekkerianus malabaricus	1150 hrs	Tomahawk & Havahart	NA	Banana-honey, crickets	6 months	N	Mundanthurai Plateau, India	Gupta, 2007

[1] Wells et al., (2004) captured a single loris 3 times in the canopy and the ground during an intensive mammal trapping survey.
[2] 70 captures, 20 individuals, were caught; 9 with noose traps and the remainder with wire mesh.
[3]13 pottos were captured 44 times, giving a rate of 1:800; two of the pottos were caught by hand.

Figure 1. Examples of the stages of collaring a Javan slow loris. Clockwise from upper left: measuring in the forest on a plastic ground cloth with all equipment ready; keeping the animal calm in a clean breathable cotton bag, washed every use; taking a saliva sample with a Salimetrics children's swab; applying the collar and ensuring the cable tie does not slip with appropriate glue, kept away from the fur with a piece of cardboard; taking a photograph with Munsell colour chart to ensure colour correction; taking standard body measurements with electronic callipers. (Photos Little Fireface Project).

Figure 2. Example of a datasheet used for the study of Javan slow loris in Indonesia for the recording of morphological data, photos, health and samples during capture.



CAPTURE DATA SHEET LFP – N. javanicus, West Java

Loris caught in the wild ☐
Loris brought by villagers ☐
If brought by villagers fill the associated form and attach it to this form
Identification band
R L
Hands

Date of trapping
Micro-Chip ID
GPS location (mark tree!)
X : GPS code
Y :
Habitat
Number/ID | Sex | Age | Name:
Capture Team
Observer, Assistant:
Trackers:
Gland secretion (dry/ moist/ exuding)
Time of capture:
Time of release:
New equipment
☐ VHF Collar……………FREQUENCY:______________ Serial number:___________
☐ Temperature logger………………Settings:______________________________
☐ Activity logger……………………..Settings:______________________________
☐ GPS tag……………………………Settings: ______________________________
Describe reaction during capture

Weight (g) ☐ with bag (100g) ☐ with collar (19 g) ☐ with actiwatch (10g) ☐ with GPS tag (1g)
Notes
Body length (neck to base of tail) | Tail (mm)
Head length (mm) | Muzzle length (mm) | Chest girth (mm) | Dark chest girth (mm)
Head width (mm) | Anogenital length (anus to base of genitals) | Neck girth (mm)
Lgth LT testicle | Lngth RT testicle | Width LT testicle | Width RT testicle | Width both testes together | Skin fold (mm)
State of vagina (clos/op/swollen) | Reproduction status (enlarged nipples, swollen/milk nipples/sexual testes)

LT | RT
Ear length (mm)
Ear width (mm)
Hand span (mm)
Hind foot span (mm)
Leg Upper (mm) Lower (mm) | Upper: Lower:
Arm Upper: Lower:

Samples
O Ectoparasites
O Gland exudates
O Saliva
O Urine
O Feces
O Pluck some hair
☐ head
☐ back
☐ belly
☐ bum

Pictures
O Face
O Profile
O Dorsal stripe
O Teeth
O Wounds

CAPTURE DATA SHEET LFP – N. javanicus, West Java

Colour of nose | Hair length (mm) Measure collected ones once back at the station
Colour of Facemask (orangish, brownish, blackish) | Width of face stripes (thick, medium, thin)

DRAW Dorsal stripe
Which vertebrate area does stripe begin and end?
Start: ☐ cervical ☐ thoracic ☐ lumbar ☐ caudal
End: ☐ cervical ☐ thoracic ☐ lumbar ☐ caudal
Thick / Jaggy / Straight?

Hair length
Head ☐ short ☐ fluffy
Shoulder ☐ short ☐ fluffy
Back leg ☐ short ☐ fluffy

DRAW Facial pattern
Examples of facial pattern
m n o
s q

Condition of ears
R L
Wounds
Describe wounds and wounds location
Other identification signs

Figure 3. Left shows a pygmy loris in a leather collar with a rubber casing; non-reflective bird bands are used on the collar to give the animal a unique colour code. Note the gloved hand of the researcher. Right shows a red slender loris in a cable tie collar, wrapped in protective plastic shrink wrap to protect the skin. In both cases, antennae were left free for a better signal; chili pepper can be applied by the manufacturer to prevent chewing. (Photos K.A.I. Nekaris)

25a. Red Light for Nocturnal Observations

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A key to studying the behaviour of lorises and pottos in both wild and captive settings is the use of an appropriate light. Here we review why red light is important and recommend lights for field usage as of 2019.

Since at least the 1950s (Southern 1955), red light has been advocated for use in nocturnal animal observations. Not only do animals perceive the red light less and engage in more normal behaviours, but the red light also preserves the human viewer's night vision. Another major disadvantage of white light is that it can dazzle the observed subject (Finley, 1959), disturbing the night vision of wildlife resulting in increased predation risk for smaller species, and disrupting natural movement or foraging behaviour (Wolf and Croft, 2012). White lights, including from on-site vehicles, have been shown to trigger a flight response in observed animals or, at the very least, dramatically alter their observed behaviour (King and King, 1994; van Geffen, *et al*., 2014; Koli and Bhatnagar, 2016). For example, the desert woodrat (*Neotoma lepida lepida*) remained largely undisturbed during red light observations while rapidly retreating to shelter during white light exposure (Thompson, 1982). A complaint of using red light is that it may not adequately illuminate the area under observation (Harvey *et al*., 2012). Despite this challenge, it is highly reflective of the eye-shine generated by the *tapetum lucidum* of nocturnal species even when the whole animal is not in focus (King and King, 1994).

For nocturnal primates, red filters on lights have been advocated since the 1960s (Charles-Dominique and Bearder, 1979). In a field study of Mysore slender lorises, any light other than red caused freezing behaviour and even prevented the animals from entering their sleep sites (Nekaris, 2001). Moreover, pygmy lorises actively look away from observers when employing white light, while appearing entirely undisturbed when viewed under red light (Starr *et al*., 2011). During a long-term study in Java of Javan slow lorises, we compared animals habituated by observers who used white light versus those who used red and found that white light highly significantly increased the amount of time that animals froze in the presence of the observer (Fig. 25a.1).

The benefit of red light use in observation is not limited to work in free- or semi-free ranging field work. Nocturnal primates housed in captive environments display overall higher activity in red light housing than blue light, going so far as to use their nest boxes significantly less in red light, which suggests an active avoidance of the blue lighting (Fuller *et al*., 2016).

Pure white light, especially from LED torches, can injure the eyes of nocturnal animals (Harvey, *et al*., 2012; Hunter, *et al*., 2012; Peng *et al*., 2012). LED usage, particularly at night, is often measured with respect to human visual sensitivities (Gaston *et al*., 2015). Consumer available LED bulbs pose little to no risk to the general public's eye health in domestic settings (Behar-Cohen *et al*., 2011). Nocturnal primates, however, have the largest eyes relative to skull size (Ross, 2000), which is associated with increased sensitivity to light (Neagu and Petraru, 2015). The rod outer-segment present in nocturnal animals collects more light energy, increasing the overall likelihood of rod and cone damage (Organisciak and Vaughan, 2010) in as little as 15-minutes after light exposure (Fuller *et al*., 1978). The sensitivity of rods in nocturnal primates is weighted to the blue end of the light spectrum, making the retinal response to both blue and white light similar but somewhat alleviated in response to red light usage both in the field and captive setting (Fuller *et al*., 2016).

Red light filters are used in other nocturnal disciplines including hunting and astronomy as a major aid for the human observer and are commercially readily available (Table 25a.1). Rods in the human eye are sensitive to light and it takes only a brief exposure to bright light for the rods to over expose. Thus even checking a notebook with a white light means up to 30 minutes are needed to regain night vision. As with primates, human rods are less sensitive to red light, meaning that our observations of nocturnal primates are much better under red light. Clearly, red light use provides less disturbed data and reduces the risk the animal may suffer from predation or altered behaviours.

Table 25a.1. Torches that come with or have optional attachable red filters that have been field tested during long-term studies of lorisiform primates. As of 2019, the Clulite options are the best at field sites with options for recharging whereas LED Lenser are best at sites where batteries are a necessity.

Model	Manufacturer Suggested Duration (hrs)	Field duration (hrs)	Rechargeable? (Internal)	Red light YN	Weight (g)	Bulb Type	Distance beam can reach (m)	GBP Price
Torches suitable for field observation								
LED Lenser H14R2	6-35	4	Y	Y	340	LED 60-1000 lm	300	153.95
LED Lenser H7R2	4-30	4-5	Y	N	165	LED 20–300 lm	160	87.95
LED Lenser SEO 7R	5-20	3-4	Y	Y	93	LED 20-220 lm	130	82.45
Clu Lite Super Spot HL13	18	12	Y	N	322	LED 3w	250	32.00
Torches suitable as an extra trail light but not to observe animals								
Petzl Tikka	240	28	Y	Y	86	LED 200 lm	60	30.00
Petzl TakTikka	60-260	37	N	Y	85	LED 250 lm	70	45.00
Black Diamond Spot	30-175	66	N	Y	88	LED 300 lm	80	35.60
Black Diamond Icon	70-175	72	Y	Y	300	LED 500 lm	100	89.01
Maglite Mini	11-32	54			118	LED 97 lm	145	15
Silva TrailSpeed	1.5-6	2	Y	N	135	LED 500 lm	80	190.29
Silva Exceed	1-20	3	Y	Y	157	LED 1500 lm	175	318
Torches suitable for spotting, filming or from a vehicle								
CluLite Fan 1 Light Fantastic	3-20	6	Y	N	460	CREE LED	500	82.50
Clulite Smartlite LiIon SM126-L1	2.25-5	3	Y	N	1350	Xenon 50w	1000	136.00
CluLite ML8 Pro Spot Torch	3-20	12	Y	N	236	CREE LED 250lm	350	62.00
LED Lenser P72 torch	50	14	N	N	175	LED 320 lm	260	69.95
Maglite 4 cell D	9	6	N	N	1043	LED 168 lm	267	28.99

Figure 25a.1 Amount of freeze behaviour showed by *Nycticebus javanicus* at Cipaganti, Java during the first six months of observation compared between animals habituated under white versus red light. Data are estimated means and standard errors extrapolated from a Generalised Linear Mixed Model with freeze behaviour as dependent variables (fitted to a Poisson loglinear function for count data), individuals as subjects and random factor, total number of observations as weight. Fixed factors considered were light (red vs white: $F_{1,34}=9.94$, $p=0.003$), period (1-3 months vs 4-6 months: $F_{1,34}=13.32$, $p=0.001$), and the interaction effect light * period ($F_{3,34}=0.27$, $p=0.608$). A sequential Bonferroni post-hoc test showed that freeze behaviour during the period 1-3 months using white light was significantly more frequent than the other light * period categories ($p<0.001$ for all pairwise comparisons